

CARNIVOROUS PLANT NEWSLETTER

Journal of the International Carnivorous Plant Society

Volume 48, No. 4

December 2019





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Journal of the International
Carnivorous Plant Society
www.carnivorousplants.org

Volume 48, Number 4
December 2019



Front Cover: A small ant (*Lasius neglectus*) attracted by the alluring glands inside a *Dionaea* trap. Photo by Siegfried R. H. Hartmeyer. Article on page 153.

Back Cover: Venus flytraps can be grown for years indoors without dormancy. Photo by John Brittnacher. Article on page 178.

Carnivorous Plant Newsletter is dedicated to spreading knowledge and news related to carnivorous plants. Reader contributions are essential for this mission to be successful. Do not hesitate to contact the editors with information about your plants, conservation projects, field trips, or noteworthy events. Advertisers should contact the editors. Views expressed in this publication are those of the authors, not the editorial staff.

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Date of effective publication of the September 2019 issue of Carnivorous Plant Newsletter: July 31, 2019.

The ICPS is the International Cultivar Registration Authority (ICRA) for the names of cultivated carnivorous plants according to the International Code of Nomenclature for Cultivated Plants. Send relevant correspondence to the ICPS, Inc.

Carnivorous Plant Newsletter is published quarterly in March, June, September, and December by the ICPS, Inc., 2121 N. California Blvd., Suite 290, Walnut Creek, CA 94596, USA. Periodicals postage paid at Walnut Creek, CA and additional mailing offices. Postmaster: Send address changes to ICPS, Inc., 2121 N. California Blvd., Suite 290, Walnut Creek, CA 94596, USA. Printed by Allen Press, Inc., 810 E. 10th Street, Lawrence, KS 66044. Logo and masthead art: Paul Milauskas. © 2019 International Carnivorous Plant Society. All rights reserved. ISSN #0190-9215

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KENNY COOGAN: ICPS EDUCATION DIRECTOR

Kenny Coogan started his career by working at two zoos and an aquarium for ten years in the education departments. He is a prolific writer, contributing over 400 printed articles to the magazines Countryside, Hobby Farms, Chickens, Backyard Poultry, and Florida Gardening. Kenny is working on his fourth TED-Ed talk. His TED-Ed video “Why are sloths so slow?” has over 3 million views and his newly published YouTube film “The wild world of carnivorous plants” is approaching 500,000 views. He is a certified middle school science and agriculture teacher. He is entering his second year of his Wipro Science Education Foundation Fellowship at University of South Florida. The fellowship provides science teachers in the Tampa Bay region with the tools they need to become more effective instructors, advance science education curriculum, and achieve greater student success.

Kenny has been keeping carnivorous plants for ten years. Last year he obtained his nursery license and sells carnivorous plants at the area’s seasonal plant sales.

His goal as the ICPS Education Director is to create infographics and a book specifically for school age children.



AN INTRODUCTION TO THE SAN DIEGO CARNIVOROUS PLANT SOCIETY

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In the heart of San Diego's beautiful and historic Balboa Park, San Diego County's carnivorous plant enthusiasts gather to exchange thoughts, ideas, and above all, plants. Each meeting, hundreds of carnivorous plants of several varieties can be found, grown by SDCPS members in San Diego's mild climate.

Established in 2013, The San Diego Carnivorous Plant Society has quickly grown to several dozen regular members of various backgrounds and experience, along with curious newcomers always stopping in for a look. With a strong start with visits by carnivorous plant luminaries such as California Carnivores' Damon Collinsworth, the SDCPS has formed a close community of like-minded enthusiasts. The bimonthly meetings consist of casual socialization among members, where carnivores are displayed by proud growers and exchanged to grow gardens, and a more formal presentation. Topics for these presentations always vary widely, from home cultivation tips, to botanical expeditions, to carnivorous plant-themed art.

The SDCPS also has a strong dedication to community outreach outside of scheduled meetings, such as reaching out to local schools for presentations and events and participating in Balboa Park public events like their annual Fiesta Botanica and Halloween Family Day, all with the goal of educating the public on the biology, cultivation, and conservation of these wonderful plants. In thanks for being its host, the SDCPS has left a permanent mark on Balboa Park in the form of a carnivorous plant bog built within the Balboa Park Botanical Garden. Established by SDCPS members' plants and maintained by the society, the garden displays the wide diversity of carnivorous plants with a goal of educating visitors on the ecology and physiology of carnivorous plants. It is always a centerpiece of visitors of the Botanical Garden.

Every summer the SDCPS holds its centerpiece event at Balboa Park, the annual SDCPS Show and Sale. This is an all-day event of botanical displays, show competitions, public plant sale, educational outreach, and children's activities. Plants sold at the show come from the personal gardens of SDCPS members, along with guest vendors such as carnivorous plant nurseries Predatory Plants and Xtream Plants. This allows for a very wide variety of plants sold that day, a sight that always surprises and delights visitors unfamiliar with the variety of carnivorous plants. SDCPS members host booths at the show with poster displays, live plant examples, presentations with plant dissections, and children's activities including themed arts and crafts and scavenger hunts, all to educate the day's visitors on the carnivorous plants they will see. This event grows every year, with the turnout in 2019 easily the biggest yet.

The SDCPS recently partnered with her sister California carnivorous plant societies, the Bay Area Carnivorous Plant Society and the Los Angeles Carnivorous Plant Society, to help organize the 2018 International Carnivorous Plant Society Conference that was held in Santa Rosa, California. Several local San Diego area members attended this conference. Members can also be found attending the LACPS bimonthly meetings, held off-sync with SDCPS meetings for easy attendance to both societies' meetings.

New members and visitors are always welcome at The San Diego Carnivorous Plant Society, so if you are ever in the area and want to talk carnivores, feel free to stop by for a visit. We love to connect with plant lovers all over the world. Updates on the ongoing activities of the San Diego Carnivorous Plant Society can be found online at sandiegocarnivorousplantsociety.com, on Facebook, and on Instagram @sdcarnivorousplantsociety.

DIONAEA TRAPS SELECTIVELY ALLOW SMALL ANIMALS TO ESCAPE

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Abstract: *Dionaea muscipula* selectively allows small animals to escape using a system of interlocking features that complement each other very efficiently. Ants of the species *Lasius neglectus* (length 3.5 mm) ran through open traps, pausing on the alluring glands along the rim of the trap moving their mouthparts over them. Analysis of videos revealed the ants primarily passed along the trap rim, over the alluring glands, but sometimes ran down to the leaf base through the trigger hairs occasionally brushing by a hair without triggering the trap, because they did not deliver the two stimuli needed to trigger trap closure.

Traps observed for four weeks were estimated from sampled observations to have had a total of about 15,000 trap visits by ants during this time period. Six ants were captured during four weeks indicating a risk of capture of about 0.04%. During this same period ten prey other than ants were captured. Visits for prey other than ants was mostly nocturnal and so low that no visits were observed during the observation period. Compared with the large number of ant visits all other prey visits were orders of magnitude fewer. The selective system that allows small animals to escape includes:

- 1) Attraction of the ants away from the trigger hairs by alluring glands.
- 2) Clear visibility of the trigger hairs to a 3.5 mm ant.
- 3) The requirement of two stimuli for triggering trap closure.
- 4) The escape allowed for small animals by openings between the marginal bristles during the slower phase of trap closure that follows the rapid snap of the trap.

Since ants are known to compose about one third of the captures by *Dionaea* in its native habitat, selection against the capture of small ants not worth the energy expenditure by the plant is an adaptive mechanism.

Introduction

There has been considerable interest in Charles Darwin's (1875) idea that *Dionaea* has a mechanism that would mainly capture moderately large insects and allow most of the small ones to escape. While doing research to test Darwin's hypothesis, entomologist Frank Morton Jones (1923) conceived of a second mechanism that would prevent the capture of small animals. The aim of this study is to reinvestigate their hypothesis and to examine in a statistically reliable manner if small animals (ants in this study) have a significantly reduced risk of being captured.

Darwin's hypothesis: The mechanism Darwin (1875) proposed for this involves the escape of prey through the openings between the projections from the edge of the leaf that remain partly open after the early rapid snap of the capture movement and during the slow closure that finally seals the trap and the fate of the prey. Darwin noted the saving in time that would result in digesting only prey that would provide sufficient nutrition and stated, "this advantage is secured by the slowly intercrossing marginal spikes, which act like the meshes of a fishing net, allowing the small and useless fry to escape" (Fig. 1).

Jones' hypothesis: Frank Morton Jones (1923) noticed that small ants were attracted to glands along the upper margins of the trap lobes. These ants moved in and out of traps without disturbing them while working their mouthparts on the marginal areas where alluring glands occur (Fig. 1). Jones proposed that small ants and other animals attracted to the glands along the leaf margin, that were too small to reach from that position to the trigger hairs, would fail to spring the trap. The result is selection of prey larger than small ants without the trap even having to bear the cost of reopening after being triggered.



Figure 1: Top: Traps directly after closure. Darwin hypothesized small prey escapes through the overlapping spikes. Bottom: Open traps with visitors. Jones, in addition to accepting Darwin's idea, also hypothesized the alluring glands guide small animals away from the triggers.

Darwin's and Jones' hypotheses are not mutually exclusive, either or both can select for prey that is sufficiently large to offset the expenditure of energy by the capture mechanism. Jones (1923) suggested that both mechanisms are involved in the selection of prey by *Dionaea*. Of course, Jones' mechanism would only act on animals attracted to the marginal glands while Darwin's mechanism would act on all prey captured.

Alluring glands: Jones (1923) observed ants attracted to areas near the rim of *Dionaea* traps and Lloyd (1942) accurately described Jones' ideas in his classic work "Carnivorous Plants", adopting the term "alluring glands" for structures in the area that attracted the ants thus making it the standard term for these structures.

The function of the trap area with alluring glands described by Frank Jones (1923) was to attract small animals, especially ants, away from the trigger hairs so that only those long enough to touch them when their mouthparts were engaged would be captured. Since about one third of the prey captured by *Dionaea* in its natural habitat are ants (Williams & Hartmeyer 2017) this function is likely to be very important.

In most papers since the 1980s the alluring glands are proposed as a lure that draws prey into the trap where it trips the trigger hairs and is captured. This is likely due to this role for the glands being described in Juniper, Robins and Joel's classic book "The Carnivorous Plants" (1989). While there clearly is an attractant produced that draws ants (Jones 1923) and flies (Williams & Hartmeyer 2017) it does not seem to draw prey into the trap from a distance.

Analysis of prey: Prey capture success by three different sized traps has been determined to have no correlation with prey size (Hutchens & Luken 2009; Luken 2019). More recent laboratory research by Davis *et al.* (2019) found larger trap size correlated with a substantial increased probability of capture. Prey mass resulted in a slightly smaller probability of successful captures. In these experiments cultivated flytraps of 5-30 mm were exposed to lab grown crickets of 7-23 mm. Since the traps were exposed to prey of moderate to large size relative to the trap and not to small animals neither Darwin's nor Jones' hypothesis was tested in the experiment.

It is unclear if the correlations observed by Hutchens and Luken (2009) and by Davis *et al.* (2019) demonstrate that smaller prey is escaping through the leaf spines and moderate sized prey is

retained. The calculations are based on prey captured. We know very little about prey that got away. Since prey capture is a relatively rare event, testing of Darwin’s hypothesis is likely to be difficult but Frank Jones’ (1923) descriptions of ants being frequently noticed on the leaves indicate that it may be possible to test his hypothesis.

The objective of this paper is to repeat Frank Jones’ observations and measure the frequency of small ants that escaped from active traps without closing them and to measure and identify the captured prey.

Methods

Dionaea muscipula in this experiment are a population of plants established more than 20 years ago in a 40 cm pot inside a garden pond in Weil am Rhein (Southwestern Germany, Fig. 2). These plants, similar to those found in the wild, are the same ones used in a previous 80-day study of prey captured by *Dionaea* in various habitats by Williams and Hartmeyer (2017).

Ants (*Lasius neglectus*, length 3.5 mm) observed in the study had established themselves in the 40 cm pot near the *Dionaea* since the 2017 Williams and Hartmeyer study. All ants and prey animals are those occurring in the garden with no manipulation by the experimenters.

Observations and video (Hartmeyer & Hartmeyer 2019) of ant visits to the traps were made. In spring 2019, the ant population settled for the first time inside a large pot with *Dionaea*, *Drosera rotundifolia*, and *Sarracenia minor* var. *okefenokeensis*, directly beside the established *Dionaea* that started to sprout again after winter dormancy. The behavior of the ants and their interaction with the active traps was observed.

Scheduled observations: Ant visits to the population of *Dionaea* traps were observed continuously during 24 ten-minute intervals spread over six days. These observation periods were made at different times of day. The time, weather, and number of open and closed traps were recorded. The prey found in reopened traps was measured and identified along with the length of the reopened traps. During the experiment the number of active traps increased from 24 to 38. The distance from the alluring glands to the nearest trigger hair was measured for traps of a range of sizes. The median length and the lengths of the largest and smallest traps were determined. Blackening or inactive looking traps were removed to keep the setup clear. However, except for



Figure 2: Garden pond population of *Dionaea*.

that, the plants were left to themselves. In addition to the removal of blackened-inactive traps new traps developed; therefore, the total of open and closed traps in the log changes. Only healthy active traps have been counted. In summer 2016, the same population of traps was documented for prey capture during 80 days (Williams & Hartmeyer 2017), which provides comparable data of the usual prey capture without ant nest beside the plants.

Photography and videography were done with an Olympus SH-2 (photos & HD-video) and a Huawei P20 Pro (4K-videos). The photographs were improved for image quality with MAGIX Photo and Graphic Designer. The original 4K-videos were edited with EDIUS Pro 9 for display details, image quality, and stabilization and then rendered to HD-format. The image stabilization was necessary because the setup is placed inside a garden pond and it was impossible to install a tripod for suitable macro shots. Therefore, the manually shot macro videos were strongly blurred. The resolution was greatly improved with the previously mentioned stabilization filter. However, no video content has been manipulated with editing software.

Results and Discussion

Observations and video of ant visits to the traps showed ants nesting in the 40 cm pot frequently moved over the plants and through the traps without being captured. The numerous ants that entered the stand of *Dionaea* in the garden pond were continuously active throughout daylight. They crawled over the plants and ran through open traps, pausing on the visibly-dry alluring glands and moving their mouthparts over them (Front Cover). Their interest in these structures was striking and is clearly visible on the shots. Analysis of the videos revealed the ants primarily passed along the trap rim, over the alluring glands. Due to the ants' small size they did not reach the trigger hairs when they passed along the rim of the trap. Even when they suddenly ran down to the leaf base through the trigger hairs, we never observed them trigger the trap. Occasionally they passed close to a trigger hair and may have brushed against it but since two stimulations of a hair or hairs within 20 sec are required to trigger closure of a trap it is very rare that this happens. When the mouthparts were on the alluring glands the 3.5 mm ants stretched only about 0.7 of the median distance to the trigger hairs closest to the rim. While they were longer than the alluring gland to trigger hair distance in the smallest trap (ratio 1.17), the ants were usually nearly horizontal to the rim of the trap and well away from trigger hairs. They did not trigger a response this way. Our observations

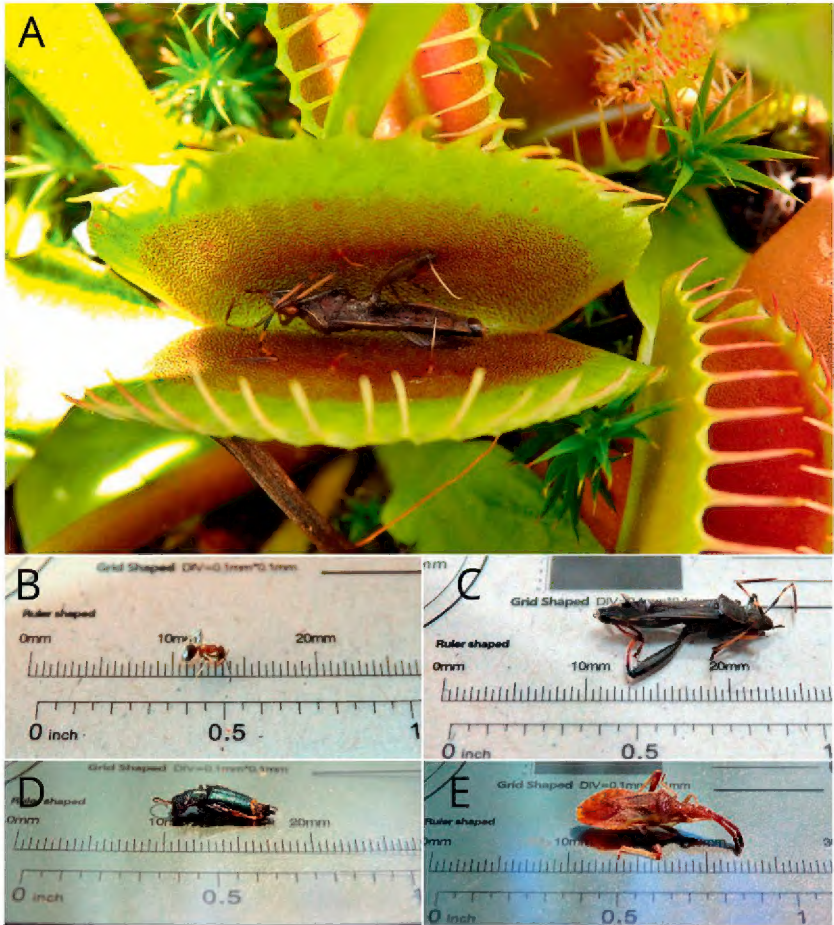


Figure 3: A&C = prey predatory bug (13 mm). B = *Lasius neglectus* (3.5 mm). D = prey beetle (7 mm). E = prey true bug (9 mm).

agree exactly with those of Jones (1923) and support his hypothesis that the alluring glands drew the small ants away from the trigger hairs and prevented their capture.

When the observations started on May 27th only three of 24 different sized active traps were closed with prey despite the heavy ant traffic. For example, in the early afternoon, 15 ant visits occurred within ten minutes, yet no captures were made that day. The first closed leaf opened in June revealing a large predatory bug (Reduviidae, Hemiptera; Fig. 3). Jones predicts that smaller animals attracted to the alluring glands will not trigger trap closure, larger animals would reach the trigger hairs and would have a high probability of being captured. Since three independent studies (Jones 1923; Lichtner & Williams 1977; Hutchens & Luken 2009) show that about one third of the prey captured by *Dionaea* in its native habitat are ants, it is clear that selection of size is important for this class of prey. Without prey size selection, almost all traps would be closed on ubiquitous small ants.

Scheduled observations of ant visits: During the 10-minute intervals all ants leaving any trap without triggering closing were counted. Adding all ants counted in 24 ten-minute intervals is equivalent to 240 minutes or four hours of precise counting, we achieved a sum of 158 escaped individuals. During this period an average of 40 ants per hour entered and left the traps without triggering them. Extrapolated to a daily period of 13 hours (counting between 9 am-10 pm) 520 problem-free trap visits daily (Fig. 4) or about 15,000 (extrapolated 14,560) during the four-week observation time. Only six ants were captured during the 4 weeks (Table 1). The risk of *Lasius neglectus* being captured is thus very low, about 0.04%. For comparison: The risk of mortality by medical malprac-

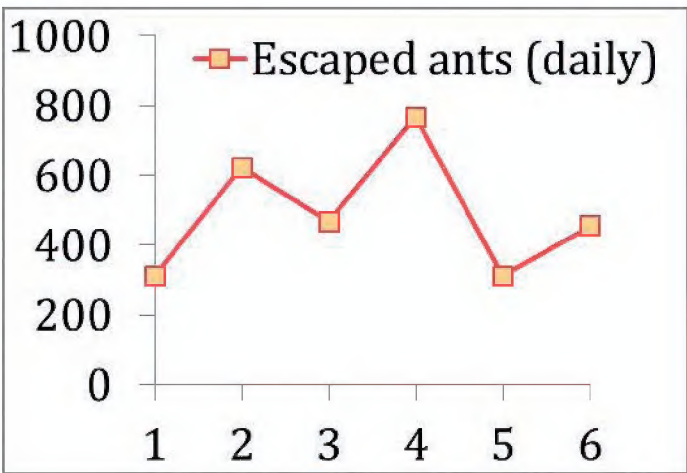


Figure 4: Ant escapes during 24 ten-minute intervals over six days, extrapolated to daily events. Average = 520 escapes daily or 14,560 in four weeks.

Table 1. Prey capture by the garden pond population of <i>Dionaea</i> over four weeks.				
Captured Prey During Four Weeks	Number Captured	Prey Length (mm)	Trap Length (mm)	Remarks
Hymenoptera (<i>Lasius neglectus</i>)	6	3.5	13-23	3 single captures. 2 ants caught at once, each carrying 4-5 mm long item. 1 ant caught together with a crab spider.
Arachnida	1	4	22	likely <i>Misumena vatia</i>
Coleoptera	5	6.5-7	18-26	Beetles, all <i>Malachius bipustulatus</i> .
Diptera	2	4 & 6.5	14 & 20	Mosquitos
Hemiptera	2	9 & 13	31 & 25	Different true bugs
Unidentified	2	3-6	2 × 20	Jelly & droppings, excluded from calculation.
Total: 16 prey animals in four weeks = 4 captures per week by 24-38 traps.				

tice for a human in a German hospital is 0.1% (BMG 2007) and thus 2.5 times higher than the risk that small ants will be captured by a *Dionaea* trap.

Prey other than ants captured during four weeks of observation (10, Table 1) consisted of five beetles (6.5 - 7 mm), two true bugs (9 & 13 mm), two mosquitos (4 & 6.5 mm) and one small crab spider (4 mm) that was captured together with an ant in one trap. All these prey animals were larger than the 3.5 mm ants, often reaching a multiple of the ant length (Fig. 5). Two ants, each carrying 4-5 mm long items of nesting material through a trap were captured in a single trap closure possibly due to their extended size. Only three ants were captured alone; however, to keep our calculation conservative we included all six captured ants, disregarding the circumstances of capture. Data on the risk of capture for prey animals other than ants is unavailable because their visits, which are mainly nocturnal, are far less frequent. Measuring their captures and escapes would require 24-hour observations for several weeks; therefore, this type of measurement has not been done. The beetles, true bugs, and mosquitos were all captured over night, when the ant traffic around the *Dionaea* paused. Compared with the large number of ant visits (maximum counted escapes in ten minutes = 21, minimum 1), all other animal visits were orders of magnitude fewer. That means even a conservative estimate of their risk of being captured is orders of magnitude higher than 0.04%. Therefore, our experiments show clearly that *Dionaea* sorts out prey of insufficient size, particularly small ants, thus increasing the chance to capture medium to large prey.

Dionaea's complex prey selection mechanism results in selection of prey of sufficient size to offset the costs of the snap trap capture mechanism. Capture of prey involves loss of resources to the plant:

- 1) The closed trap has reduced photosynthesis due to the change in orientation of the leaf to the sun, decreased flow of CO₂ and changes in metabolism (Pavlovic 2010).
- 2) Energy is used in closing and reopening the trap.
- 3) Energy is expended in digesting prey.
- 4) Traps can make only 3 to 4 closures; each closure is slower than the previous one. The snap of a trap is therefore a limited resource (Brown 1916; Davis *et al.* 2019).

The advantages of prey selection relate to the relative costs in capturing prey of different size. Small prey probably have an energy expense out of proportion to any advantage gained. While the



Figure 5: 1) The alluring glands draw ants away from the triggers. 2) The trigger hairs are large enough to be visible to ants crossing the trap. 3) One accidental touching of a trigger hair does not close the trap. 4) Even if the trap closes, gaps between the bristles allow small prey to escape.

advantages of carnivory in *Dionaea* are related to procuring nutrients rather than energy the energy expenditure must still be worth the gain in the overall budget of the plant.

The *Dionaea* prey selection mechanism is a system of at least four different interlocking features which complement each other very efficiently.

- 1) The dry alluring glands attract some animals, such as ants and flies, away from the trigger hairs. Of the animals attracted only those long enough to reach from the alluring glands to the trigger hairs have a high probability of being captured (Jones' mechanism). These visitors often pause and move their mouthparts over these glands which occur near the marginal spikes and they leave and enter the traps often through the spaces between the spikes or at the space at the petiole without crossing the dangerous trigger zone (see Hartmeyer & Hartmeyer 2019 video and Fig. 5).
- 2) Frequently, ants suddenly walk down to the leaf base and pass the dangerous trigger hair region. However, ants are unlikely to run headlong into visible obstacles. With a length of approximately two millimeters, the trigger hairs should be clearly visible for a 3.5 mm sized ant (see Hartmeyer & Hartmeyer 2019 video and Fig. 5).
- 3) Even if a leg or antenna touches a trigger hair accidentally nothing happens, the traps remain open. *Dionaea* responds within about 20 seconds for a second contact. Without it, the trap remains open and the memory of the first stimulus fades and another two stimuli are required for closure.
- 4) After stimulation by prey a *Dionaea* trap will rapidly snap closed but the closure is not complete. A gap remains between the lobes for several minutes. During this period the escape of large prey is barred by the marginal spikes, which cross over each other along the open edge of the trap. Animals that are small enough have a chance to escape between the marginal spikes (Darwin's mechanism). This mechanism will allow the escape of small animals regardless of whether or not they are attracted to the alluring glands.

The interaction of all these features enables small ants to visit *Dionaea* traps with a risk of only 0.04% of being captured. This amazingly small risk suggests that *Dionaea* clearly has complex mechanisms that prevent the capture of small ants. Other small prey, not attracted to alluring glands, can still escape by mechanisms 2, 3, and 4 if they are agile enough. However, observations that would prove this in detail have yet to be made.

Previous experiments on prey selection have been inspired by Darwin's suggestion that small potential prey escapes during the later phase of closure when gaps between the marginal spines provide an escape route. This is an ingenious idea and also attracts attention because it was the famous Charles Darwin who suggested it. Three of the studies based their conclusions on measurements of the prey captured in the field. Darwin (1875) believed that his limited observations supported his hypothesis and Jones (1923) felt his observations supported both Darwin's and his hypothesis although the results were not definitive. Hutchens and Luken (2009) found no significant correlation between prey size and trap size and concluded that, attractive as Darwin's hypothesis was, the evidence does not support it. All of these studies looked only at prey captured and had no measurements of small animals that escaped. If either hypothesis is correct it must be demonstrated that small potential prey has a much larger chance of escaping than large prey so none of the studies is conclusive. Davis *et al.* (2019) did measure the probability that crickets of a range of sizes would be captured under laboratory conditions and therefore had a chance of testing Darwin's hypothesis when crickets are the prey. However, their data shows smaller crickets have a higher probability of being captured than larger ones, exactly the reverse of what would be expected if Darwin's hypothesis was correct. However, since the crickets range between 7-23 mm while the traps range from

5-30 mm (except for one small trap) the crickets should be considered medium to large prey. They did not investigate potential prey small enough to test Darwin's hypothesis rigorously.

Jones' mechanism of attraction of ants away from the trigger hairs toward alluring glands on the rim of the trap allowing them to escape is effective. Darwin's mechanism of allowing small prey to escape through openings between the marginal spines along the lobes of the trap during the slow final phase of closure probably also works, but more observations and experiments are needed to confirm this in detail. An experiment like that of Davis *et al.* (2019), where the probability of capture of medium sized and large prey but with small potential prey like the ants in our experiments included is probably the best way to test Darwin's hypothesis. Considering the amazing fact that during our experiment, which was done over four weeks, only six of 15,000 ant visits triggered a closing makes it very likely that such an effective sorting out of small animals is based, not on just one mechanism, but on a sophisticated system of several interlocking features that include the mechanisms of Jones (1923) and Darwin (1875).

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QUINONE PATTERNS AND IDENTIFICATION OF JAPANESE SPIDER LEG SUNDEWS (*DROSERA* SECT. *ARACHNOPUS*)

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Keywords: *Drosera*, Phytochemistry, Naphthoquinones, Chemotaxonomy.

Introduction

The Japanese sundews that have previously been collectively called *D. indica* L. (especially after the influential Flora of Japan, Ohwi 1965:492) are being split up. Various authors (e.g., Watanabe 2013) are trying to re-establish or create one or two additional species assumed endemic to Japan, following the contemporary trend to recognize a number of segregate taxa in *Drosera* sect. *Arachnopus* (Greek for “spider leg”), the group that contains *D. indica*. Previous research (Schlauer *et al.* 2017, 2018, 2019) has demonstrated a rather unexpected diversity in the naphthoquinones that are characteristic for the different species now recognized in this group. While a few Australian species contain ramentaceone (7-methyljuglone), most contain the regio-isomer plumbagin (2-methyljuglone). So far, only *D. indica* s.str. (accessions from Asia and Africa have been investigated) contains both isomers in the same plant. The morphological diversity found in Japan thus prompted a chemical investigation, the results of which are reported here.

Materials and methods

Plants of two different accessions informally named “*D. makinoi*” (with white flowers, seeds with almost isodiametric testa cells; from Tochigi and cultivated by a grower in Kanto district, Japan) and “*D. toyoakensis*” (pink flowers, seeds with longitudinally compressed testa cells; from Toyohashi and cultivated by a grower in Kansai district, Japan) were raised from seed obtained from Koji Kondo, Kobe, Japan. The methods for cultivation, extraction, separation, and detection of quinones were the same as detailed previously (Schlauer *et al.* 2018).

Results

Naphthoquinones were detected in both samples. While the pink flowered taxon contained only plumbagin, the white flowered one contained both plumbagin and ramentaceone.

Discussion

The pink flowered plant corresponds in all morphological aspects to *D. serpens* Planch. (petiole present, emergences of insect abdomen, bulls head and bowler-hat types, seed testa cells longitudinally compressed). And, as also the characteristic quinone plumbagin is found in the Japanese plant as well as in several previously investigated samples of *D. serpens* (from Asia and Australia), there remains little reason to separate the Japanese plants from *D. serpens*. The reported localities near the

towns of Toyoake and Toyohashi (both in Aichi Prefecture, Japan) are thus new records for *D. serpens*. *D. indica* f. *rosea* Makino (1905:24) is probably a synonym, its type locality (“Takashi-mura, Prov. Mikawa”, possibly in the Toyohashi area) and another locality given by Makino (“Takatori-mura, Aomi-gori”) are both in the same part of Aichi Prefecture.

The white flowered plant is somewhat more difficult to interpret because in emergence morphology (Fig. 1) it coincides with *D. serpens*, while flowers, seeds, and the quinone pattern are more similar to *D. indica*. The plants evidently grow true from seeds, so the plant is fertile and probably not an F1 hybrid. As it seems possible to differentiate it from typical *D. indica* by its indumentum, recognition as a separate taxon may be justified. If it is derived from hybrids between *D. indica* and *D. serpens* (the hypothesis proposed here) and if the assumed parents are separated at species rank, it cannot be classified as an infraspecific entity under either (as originally done in *D. indica* f. *albiflora* Makino, 1905:24; type from Kazusa Prov., now Chiba Pref., Japan). The first validly published name at an appropriate rank would indeed appear to be *D. makinoi* Masam. (Masamune 1935:11, type from Simotuke/Shimotsuke Prov., now Tochigi Pref., Japan). Considering the large area of geographic overlap of the putative parent species, it is not necessarily confined to Japan. Future

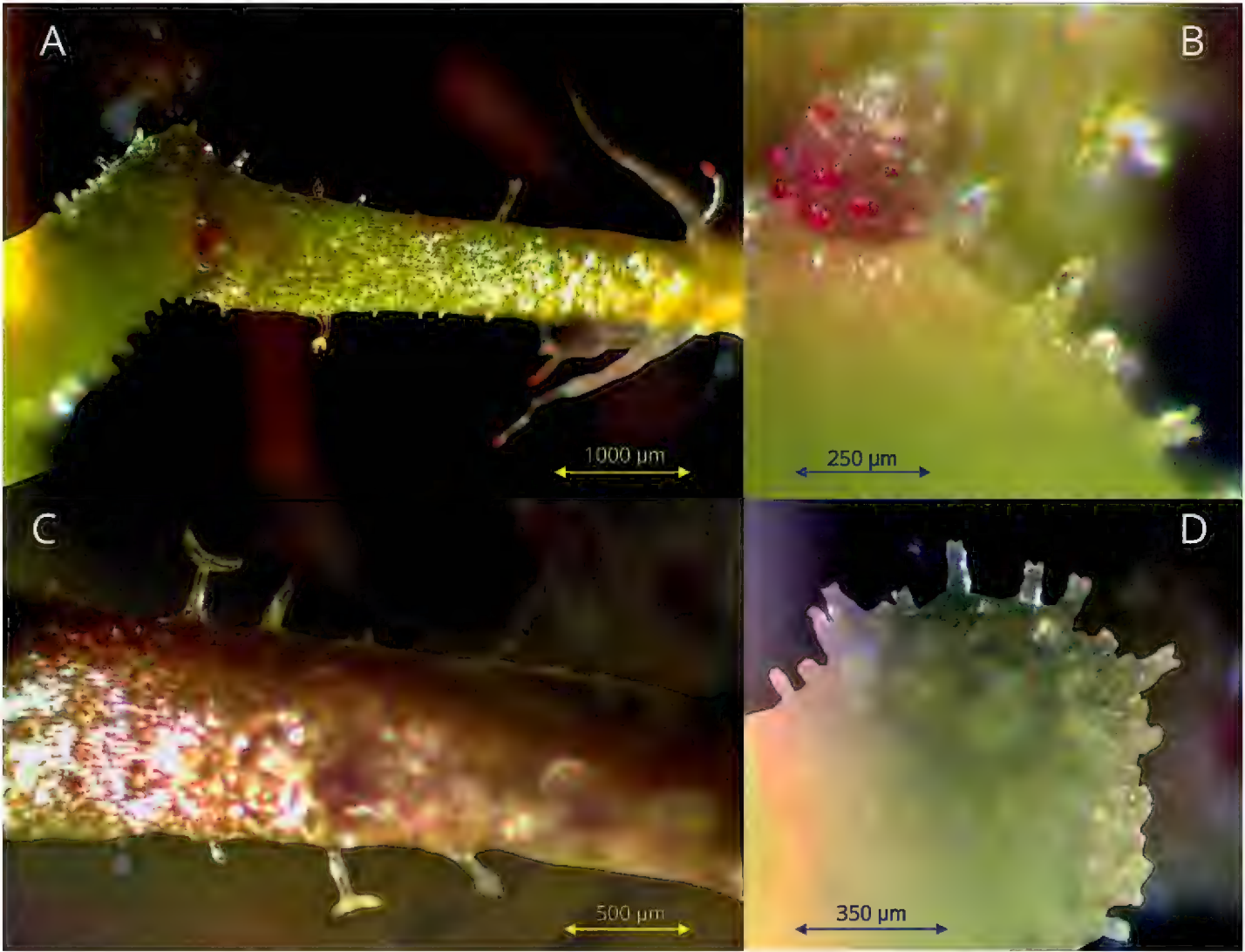


Figure 1: Indumentum of the white flowered sundew taxon assigned to *Drosera makinoi*. A: leaf base showing petiole with bulls head emergences, double-tipped-cap emergences and (small) insect abdomen emergence; B: bud in leaf axil dotted with red stipitate glands; C: detail of petiole with bulls head emergences, double-tipped-cap emergences and insect abdomen emergence; D: young expanding leaf with double-tipped-cap emergences on lower (abaxial) surface. Cultivated plants, photos by Siegfried R.H. Hartmeyer.

floristic (and phylogenetic) research in eastern Asia should take this into account. If all plants identified as *D. indica* f. *albiflora* in the last revision (Komiya & Shibata 1994) were indeed *D. makinoi*, there would not remain a single record of *D. indica* s. str. from Japan (because all others refer to *D. serpens*), which would entirely overturn the previous concept (Ohwi 1965) that had *D. indica* as the sole representative of *D. sect. Arachnopus* in Japan.

Acknowledgements: We would like to express our sincere gratitude to Koji Kondo, Kobe, Japan, for providing the seeds of the plants investigated in this study and for valuable locality and literature information, to Tian-Chuan Hsu and Jer-Ming Hu, both Taipei, Taiwan, for tracing the type of *D. makinoi* (“Hab. Isokawa, oppido Hongo, Simotuke”, Oct. 5, 1930 [not “Oct. 15, 1930”, as printed in the original description], Suzuki-Tokio s.n. (TAI051807)), and to Andreas Fleischmann, Munich, Germany, for correcting the manuscript.

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DROSERA IN THE WESTERN USA—AN UPDATE

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Keywords: Taxonomy: *Drosera*, western USA, *Drosera anglica*, *Drosera intermedia*, *Drosera linearis*, *Drosera* × *obovata*, *Drosera rotundifolia*, *Drosera* × *woodii*.

One of my primary research activities focuses on carnivorous plants in the US states of Montana, Wyoming, Colorado, New Mexico, and westward (but excluding Alaska). Throughout most of this range, the only native species of *Drosera* are *Drosera rotundifolia* L., *Drosera anglica* Huds., and the hybrid *Drosera* × *obovata* Mert. & Koch. These species are well-known to readers of this journal, and need no further description. In this region, these plants quite often occur in sites that are widely separated, and in some states these taxa are of considerable rarity (for example, in Colorado *D. rotundifolia* and *D. anglica* are known at only 4 and 1 sites, respectively; Wolf *et al.* 2006).

In addition to these species, there have been reports of two additional native *Drosera*—*Drosera intermedia* Hayne in northern Idaho (and perhaps adjacent eastern Washington), and *Drosera linearis* Goldie in Montana.

Drosera intermedia was reported in Boundary County, Idaho by Bursik (1993), as growing with *Drosera anglica*, but being quite different in form. Additional populations were subsequently found in Custer County, Idaho (L. Kinter, pers. comm. 2017). Of course, the closest other *Drosera intermedia* plants were on the other side of the continent, so this was considered a find of great interest, and the plants were treated as of special significance.

A few years ago, I decided I needed to visit and assess these plants. Because of my long history of productive engagement with regional staff, I was granted permission to visit the sites, even though the sites were considered potentially quite sensitive. In fact, agency staff even gave me supplemental information on additional *Drosera intermedia* sites I could visit. There was only one catch—grizzly bear activity had been so high in the area that in some cases I could only go accompanied by experienced staff. Of course, I welcomed this company, as it ensured that I would reach the exact, correct sites.

In late July 2017, I visited the site in Boundary County where the putative *Drosera intermedia* were first detected. In this case, my wife (Beth Salvia) and I travelled unescorted. The hike was moderately strenuous. This hike (and subsequent plant search) was somewhat stressful because of the grizzly bear issue. We were both armed with cans of bear spray (which is a more effective bear deterrent than firearms), but while we saw plenty of scat, no bears were observed. We did have the good fortune of encountering a mass-flowering of *Xerophyllum tenax*—ironically called beargrass (Fig. 1).

At the site, the candidate plants were easily detected. They did look strange—they grew in clumped masses



Figure 1: A mass flowering of *Xerophyllum tenax* in Boundary County, Idaho.



Figure 2: Clumping *Drosera anglica* at the original “*Drosera intermedia*” discovery site in Boundary County.

with small leaves (Fig. 2). However, being experienced with the large variation of *Drosera anglica* as it occurs across the western USA, I quickly realized these were simply *Drosera anglica*. Viable seed were observed, as was seedling recruitment, so it was clear these plants were not sterile hybrids.

As an aside, I note that there are a number of ways to distinguish *Drosera intermedia* from *Drosera anglica*:

- 1) The leaves of *Drosera intermedia* are held at a wide of angles around the rosette tip—from nearly vertical to horizontal—while those in *Drosera anglica* are mostly erect.
- 2) The inflorescence of *Drosera intermedia* emerges (often horizontally) from the side of the inflorescence, while the inflorescence of *Drosera anglica* is more erect.
- 3) Finally, and most reliably, the seed coats are completely different. The seeds of *Drosera intermedia* are covered with papillae (bubble-like lumps), while the seeds of *Drosera anglica* are covered with small, shiny, lens-like surfaces.

The clumpy plants matched *Drosera anglica* on all counts.

Over the next few days, Beth and I were kindly escorted to a few other sites in Idaho and Washington (Pend Oreille County), where more “*Drosera intermedia*” plants were reported. In all cases, the plants we found were simply variations of *Drosera anglica*. Why were these being reported as *Drosera intermedia*? Because the regional botanists had been told that an uncommon sundew was in the area, and any aberrant *Drosera* tended to be identified as *Drosera intermedia*. However, the truth is that they were simply seeing *Drosera anglica* being strange, as it occasionally is.

Some information for the hungry horticulturists reading this article. I have grown many different *Drosera anglica* forms—I saw nothing of horticultural significance in these plants. Furthermore, they live at high elevations (1460 m) in the mountains of Idaho, and probably spend most of the year as hibernacula. These are interesting plants, as are all carnivorous plants, but not of horticultural value.

Incidentally, while we saw black bears on our excursions, we were relieved to not have any encounters with grizzlies.

Several days later, we met with other field workers in Custer County, Idaho, to look for more “*Drosera intermedia*” populations. Once again, we found *Drosera anglica*—and at these sites there was really nothing out of the ordinary with these plants. I am guessing that, perhaps some atypically harsh year, someone was surveying these sites and encountered some plants in an abnormal growth state. Whatever the back story might have been, the Custer County plants were mundane *Drosera anglica*.

If my conclusions about their “rare” *Drosera* was not what they wanted to hear, my agency hosts didn’t show it. In fact, in some ways they were relieved because they could now tell the occasional



Figure 3: (left): Tiny *Drosera anglica* in Valley County, Idaho—each plant is only about 2-3 cm in diameter; (right) nearly anthocyanin-free *Drosera anglica* in Custer County, Idaho.

carnivorous plant growers that contacted them, that these “rare” plants were really just common after all. (Many carnivorous plant horticulturists had asked them about the plants, to learn if they could collect them.)

Unrelated to these “*Drosera intermedia*” sites, Beth and I did happen to see some interesting variation in *Drosera anglica* at other sites in Idaho. At a very remote site in Valley County (that I had visited several years before) we re-observed plants that were only a few cm across at maturity (Fig. 3 left). Meanwhile, in Custer County, we observed a population which included clumps of extremely lightly pigmented plants—almost completely anthocyanin-free (Fig. 3 right). They were growing in a very wet fen, but within meters of normally pigmented plants that appeared to be growing in identical conditions.

Having put to rest, to my satisfaction, the tale of native *Drosera intermedia* in the western USA, we travelled eastward to central Montana. The mountains of Montana are fascinating to me for their populations of *Drosera linearis*. This species is primarily found in the Great Lakes region of the USA and Canada, and sites eastward (Rice *et al.* 2017). However, there are a few sites in Canada in Alberta (and possibly Manitoba, Saskatchewan, and even British Columbia), extending the range of the plant far to the west (Rice 2019).

Flashback more than a decade

Back around 2005, while conducting background research for my first carnivorous plant book, I encountered references to *Drosera linearis* in Montana—Montana! I was astonished, but was not able to schedule a trip to see them until 2006. Beth and I had been planning a long vacation in Glacier National Park, so we set aside a few more days for an ancillary expedition to look for these “*Drosera linearis*.”

By that time, I had used research tools at my disposal to determine exactly where these plants occurred. I contacted Montana Natural Heritage Program, and chatted with staff responsible for overseeing the sites. I verified that a visit would not interfere with existing research programs or conservation efforts.

The day of our *Drosera linearis* trip began poorly. It was late fall, and an early snowstorm had struck the day before. We had to drive several hours to reach the trailhead, and 15-45 cm of snow covered the ground. During most of the drive, everything was covered by deep snow. But amazingly, when we reached the trailhead, we found that our area had somehow spared the snow, and the ground was clear.



Figure 4: (left): *Drosera linearis* fen, viewed through a veil of dead, standing trees; (right) *Drosera linearis* hibernacula in Montana.

This was black bear, grizzly bear, and mountain lion habitat. Topping off our list of hazards, hunters on horseback were common—all day long the mountain silence was punctuated by the crack!-crack!-crack! of long guns being fired. With these dangers present, Beth and I adopted a strategy so we wouldn't be mistaken for animals by predators or overly-weaponized humans. If we ever found something worth hunching over—a plant, insect, fungus—only one of us was allowed to be bent over at a time; the other person had to be standing tall. We did have a close encounter with a few black bears trotting down the trail towards us, but once they saw us, they retreated into the bush at full run.

The hike from trailhead to fen complex was arduous, but we were rewarded with spectacular views of gray owls. All around the trees had been burned by a recent forest fire, which had been so hot that the trees were killed, but not hot enough to burn them to the ground. It was eerie walking among so many dead giants. After a few hours of navigating the network of trails, we reached our site—a complex of clearings in the woods, some of which were fens with open water. Because of our slow progress, we only had time to explore one of the fens. It looked like an open, saturated clearing, but the mucky ground undulated underfoot (Fig. 4 left). It was much like a quaking *Sphagnum* bog, but the surface consisted of organic muck, apparently bound by the sedges and other emergents. And there—on the delicate floating mucky surface—we saw small, globular *Drosera* hibernacula with dead, dried leaves (Fig. 4 right). Despite their dried state, we had clearly and unquestionably found *Drosera linearis*! My first view of the species in the wild! Surely less than optimal, but a spotting nonetheless!

Flashback over...we now return to the current day

During our 2017 trip to Idaho, another journey to see *Drosera linearis* was essentially compulsory. This time we would be there during the growing season, so I hoped to see more than just hibernacula. We dedicated a few long days for this trip. I hoped to survey all 15 or 20 clearings that I could see on my aerial photos of the region.

When we arrived, I discovered a new hazard—it was an intense fire season and a crown fire in the neighboring mountain peak had a giant smoke plume making it look like an apocalyptic volcano. I chatted with burn crew, and was relieved to learn that access to my desired target sites was not being impacted, at least not yet. However, if the wind changed, we had to be ready to evacuate at a run!

On the first day on the trail, Beth and I headed out early. Our hike was made easier by our prior experience in the region, and there was less uncertainty in our route. We stopped only a few times to



Figure 5: *Drosera linearis* in Montana.

appreciate the plants and animals along the way, it would have been nice to linger, but I was insufferably anxious to study the *Drosera*.

As before, we reached a point where our path got as close as it would to our destination fen—at that point we had to bushwhack cross country. The last time (in 2006) this wasn't much of an issue—but now, the travel was considerably different. In 2006, we walked easily in an eerie forest of fire-killed, but standing trees. Now all the trees had fallen over—in random directions—so the ground was piled chest-high with crisscrossed trunks and branches. Our travel slowed to a snail's pace, as there was no simple way to traverse the land. Literally, our progress was reduced to a crawl as we negotiated the sometimes sturdy, sometimes decaying, overlapping fallen trees. Falling off the branches potentially meant falling a meter or more to the ground, and if we fell inelegantly, we might easily snap a leg on the way down. This was not what we expected, but it was what we had to do.

After a tediously long crawl over the landscape-level tree pile, we reached the open fen at last! The *Drosera linearis* plants were in full growth, mostly past flowering and in fruit (Fig. 5). Interestingly, the plants were spaced apart, unlike other *Drosera* which can readily be found in clumps or dense patches.

After exploring this site (and finding no other species present), we strategized. We had many other sites in the area we wanted to visit, but progress over the downed trees was glacially slow. Based upon our aerial maps, we found connecting corridors from site to site, which normally I would have avoided because they tend to be choked with vegetation, but in this case, they were our best options.

The rest of the day we investigated clearing after clearing, just to discover that they were all too dry, or were open water sites with no floating muck mat. A great amount of energy was spent to obtain negative results! Ultimately, it was time to leave the fens. The long, laborious crawl over downed trees was unattractive but unavoidable. It took even longer than before because we were

running out of energy—but in time we finally reached the path and rested and rehydrated before the long hike home.

About only one hour into our return hike, as we passed through a particularly dense stretch of forest, Beth (who was leading) stopped without warning, and I nearly plowed into her. “Bear! Bear!” Yes—there off to the left about 50 meters away, was a huge, shaggy brown beast larger than any bear I had ever before seen. It was busy browsing with its back to us, but as it fed it turned its head enough to one side so I could clearly see the characteristic field marks of a grizzly!

I started talking nonstop babbling sentences—quietly at first, getting progressively louder, to alert the bear as to our presence in as much of a non-alarming way as possible. And indeed—the bear suddenly turned its head in our direction, locked eyes, then exploded away from us in high speed travel over the downed trees. It was both fabulous and appalling to see how effortlessly this massive animal raced, like a giant furry hovercraft, over terrain that was so challenging for us.

The bear gone, and our veins filled with a heady blend of adrenaline, we hiked back to the car at an astonishing pace. I remember very little of that part of the trip.

The next day we again drove to the trailhead. The wind had changed, and parts of our drive were through heavy smoke from the nearby fires. This was not a happy development, as it meant the fires could be moving in our direction.

We hiked back to the complex of fens and clearings. This time, chastened by the discovery of how slow our bushwhack travel was, I set my goals for just one site. (I had spent the evening after our first day looking at the aerial maps, and observed that only one additional site resembled the mucky site where we found *Drosera* on our first day.) Unfortunately, it involved an even-longer cross-country foray, this time over terrain that also had significant variations in topography. It was also not too far from where we saw the grizzly. Needless to say, it was a challenge to our endurance as well as our nerves, but at last we reached our site—a large grassy lake. Scanning the area with our binoculars, I was able to pick out a large area where the same vegetation grew as at the *Drosera* site on the previous day. That was our target. We followed the lake edge until I found an approach to the muck mat that didn’t require too much wading. We tucked our pants into our socks to block any leeches, and made our way out to the site. The muck mat was very delicate, and Beth and I made sure to keep our distance from each other so at no point did the muck have to support both our weights. Even so, I could see that our presence was causing some damage, which we tried to minimize.

Drosera!

Yes, we were successful once again. *Drosera linearis* plants were visible growing on the muck, this time with a significantly higher density than at the first site.

As I examined the plants, I noticed something interesting. There were *Drosera anglica* plants mixed in with the *Drosera linearis*! This was exciting to me, as I had seen some references to *D. anglica* in this region. But then, much to my surprise, I observed that the fruit of these plants were sterile—there were no seed being produced. Again and again I examined the plants and found that these *Drosera* were bearing no seed—they were sterile! Furthermore, there were two morphs of these sterile plants—a long leafed form that looked like typical *Drosera anglica* (Fig. 6), and a shorter-leafed form that looked more like *Drosera rotundifolia* (Fig. 7). The inflorescences of both were much taller than the inflorescences of *Drosera linearis*.

So what were these plants? If I were in California, in areas where both *Drosera anglica* and *Drosera rotundifolia* can be found, I would have concluded they were *Drosera* × *obovata* plants. If these were *Drosera* × *obovata*, it would represent a new taxon for the state. Since neither parent was visible at the site, this was a difficult pill to swallow.



Figure 6: Long-leaved *Drosera x woodii* in Montana. *Drosera linearis* is visible to the right.



Figure 7: Short-leaved *Drosera x woodii* in Montana.

Another possibility is that these were the hybrid *Drosera linearis* × *rotundifolia* (that is, *Drosera* × *woodii*). This seemed more likely, since it would only require one missing parent—the *Drosera rotundifolia*. Perhaps a more comprehensive survey of the region would reveal this species. Even if not, it might have occurred at the site in the past, only to perish. I have found places in California where *Drosera* × *obovata* was present, but only one of the parent species occurs. So my current working hypothesis is that the hybrid plants at this site are two strains of *Drosera* × *woodii*. This is extremely exciting, since the only other examples of this hybrid are found far to the east, in the Great Lakes region (Rice *et al.* 2017).

As much as I wanted to collect herbarium specimens for this new find of scientific interest, I did not have prior permission from the agency overseeing the site, so I documented the plants with photographs and videography only. Upon my return home, I found that these hybrids had been collected from the site, although identified incorrectly as *Drosera anglica*—so science has been served!

I suspect that some collectors out there would like to grow these plants, but they are not in cultivation. I do not know how they would respond to cultivation—are they as difficult to grow as *Drosera linearis*? Is there much point, since they look essentially identical to various strains of *Drosera anglica* or *Drosera* × *obovata*? Is it critical to replicate the chemistry of the muck? Would it be possible to replicate the long months of Montana winter? In any event, visitation to the site is extremely challenging, with many obstacles.

That said, these plants would be wonderful subjects for further study. However, I must note that our short trip resulted in noticeable damage to the muck-mat. I hope that future scientists will do what they can to mitigate damage to this fascinating site.

In closing, I note that I have created a few videos about this site that I have posted on my YouTube channel. Also, Joyce Ferguson has written a wonderfully entertaining poem about this site (crafted when it was thought the sites contained *Drosera anglica* and *Drosera linearis*, instead of *Drosera* × *woodii* and *Drosera linearis*). Enjoy!

Specimens of all the “*Drosera intermedia*” were herborized for future researchers. Appropriate permissions were obtained prior to making collections.

Acknowledgements: I would like to thank our contacts in Idaho and Montana who either accompanied us on our trips, or provided data support: Braden Burkholder (Montana Natural Heritage Program), Jennifer Costich-Thompson (US Forest Service), Justina Dumont (US Forest Service), Lynn Kinter (Idaho Natural Heritage Program), Andrea Pipp (Montana Natural Heritage Program), Steve Shelly (US Forest Service), Debarah Taylor (US Forest Service).

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ODE TO THE SUNDEW

By Joyce Ferguson 2/19/2019

Oh, Sundew, pretty Sundew, I've known you just a while.
But you've been round for centuries, sipping insect bile.
T'was quite a trek to find you. But I had lots of fun.
I didn't see your picture, before my search begun.

Were you small and short, or tall and slender built?
What color were your petals? Were you plain or brightly gilt?
Well I found you with my friends, growing in the fen,
A place of carbonate acid. In the moss you made your den.

Bouncing on your fen moss and your sphagnum as it floats.
Was a joyful time of laughter as I sought you with bright folks.
Your distribution, circumboreal in the temperate zone.
Who would have thought you clever, to eat on insect bones?

How beautiful you are, at least in summer's sun.
You glisten as the diamonds. Is your nation like the Huns?
Your family, Droseraceae. Your genus *anglica*.
Near your side your cousin, *linearis* all in bud.

Oh, *Anglica*, some call you, the English Great Sundew.
But *Linearis*, *Slenderleaf*, is what the blokes named you.
All North American *Droseras* have 20 chromosomes,
But *Anglica*, you're different, with 40 that you own.

Some error in meiosis, apparently occurred,
At creation of your pollen and your ovule, you were stirred.
And from *Rotundifolia*, and *Linearis* breeds,
Anglica, you're fertile, through amphidiploidy.

Small autogamous creature, you do your own self mate.
Described to us by Hudson, in 1778.
A basal rosette at your feet, just four inches high,
Your peduncle, maybe eight, reaches to the sky.

And underneath your skirt, some tiny little corms.
Also, you're perennial, once you have been born.
Your leaves are red and green, of the hue of Christmas Grinch.
But solid red they turn, if they become sun drenched.

Your laminae may grow to be, slightly longer than an inch,
But your petioles are longer, and your rosette rather dense.
Your stalk it holds a flower, and often up to five.
But each one gets its turn to be, admired opened wide.

Your petal gown is white, and very, very thin,
Perhaps an half inch round about. Its beauty makes me grin.
Ringing 'bout your pistil are five stamen, yellow tipped.
From its top three styles, 2-lobed and purple dipped.

Oh, Anglica your laminae are rather kind of round.
But Linearis you've decided slender leaves profound.
The gorgeous part about you is your sparkle and your shine.
For you look as though the morning dew, all day about you binds.

On the edges of your laminae, you grow a hundred treats.
And sometimes on your plane sides, a shorter similar sweet.
Short, little lollipops, for all the fairies' joy.
A clear and viscous droplet tops, each one as a coy.

Yes, you're a devilish creature, or shall I say quite smart?
A lure you made to capture, any prey that round you darts.
These tentacles of yours, stalked mucilaginous glands.
Are like a thousand sticky fingers on numerous little hands.

And when the Northern Bluet, or other dragonflies,
Smell your tempting perfume, on you they soon will die.
The moment that they touch you, you grab them tight and curl,
Your thigmotropic motion, winds up, like hair upon a girl.

In fifteen little minutes you asphyxiate their spiracles.
But this your nutrient soup, is your blessed God made miracle.
A fascinating work, ingenious by design,
Just so you've got some nitrogen. God to you is kind.

Before the winter comes, and your wetland home is ice,
Your life blood's well protected, 'cause you curl your leaves up tight.
You knot them at the ground, and surround your resting bud,
Then lie dormant until spring, in your hibernacula.

Your antitussive properties help people with dry coughs.
But now-a-days, such medicine, most folks would only scoff.
If it weren't a lot of work, I might try to make some dye,
From your red and vibrant color, I would grind you, but you'd cry.

And I'd rather see your glory, and leap from float to float.
Than to haul through all that jackstraw, a fine and slender boat.
So, rest assured your glistening will carry on quite marvelous.
And your future progeny, will ever be carnivorous.

STOP SHARING INFORMATION, DAMMIT! YOU'RE NOT HELPING!

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Keywords: Poaching, field observations.

Introduction

Friend and colleague Leo Song always had, in his emails, a sig file tag line that read, “Knowledge is not knowledge unless it is shared.” He referred to it as his tao. It’s an admirable core value—one that we would no doubt benefit to follow more than we do. But there are times....there are times it doesn’t work!

The problem is this. There are many people visiting carnivorous plant sites. It’s a wonderful experience—both scientifically fascinating and spiritually recharging. A natural consequence is that they want to share news of what they have done. Sadly, this can have very unfortunate consequences. It can both excite poachers and tell them exactly where to go (Figs. 1–3).

Here we are, in 2019, and unquestionably, the poachers are still very, very active.

Below I give five steps that we can take to contribute to a solution, and not be part of the problem. Most of what I am going to say below applies to my experiences in the USA, but I bet that the lessons are exportable across much of the planet. I anticipate some of you might think I could be giving guidance to would-be poachers, but I assure you—they already know everything I describe below. Don’t fool yourself by thinking otherwise.

1) Be VERY careful of sharing site location

I have had the privilege of visiting carnivorous plant sites scattered across the continent. While many are sites I found myself, I’ve also been told about others by countless friends—including but not limited to Amorim, Barnes, Brittnacher, Casper, Collingsworth, Determann, Evans, Fleischmann, Fowler, Galloway, Gibson, Hartwell, Howlett, Hummer, Jorge, Lowrie, Mann, Mazrimas, Mazur, McPherson, Miller, Novak, Pagoulatos, Prothero, Rondeau, Schlauer, Schnell, Todd, Tryon,



Figure 1: Left: What used to be *Sarracenia flava* (Pender County, North Carolina), shortly after its location was posted on iNaturalist. Right: This dead-end end road was lined with a population of *Sarracenia flava* var. *atropurpurea*. This is what remains (Brunswick County, North Carolina). Photos: Tyler Gramley.



Figure 2: Two images of the same clump of *Sarracenia purpurea* (Brunswick County, North Carolina), separated by 33 hours. Photos: Tyler Gramley.

Yin, Zerr, and Ziemer; and then of course all my colleagues in academia, NGOs, State, and Federal agencies.

Since I have some prominence, I am frequently contacted by people I barely know, and am asked about sites to visit. Shouldn't I pay forward the kindness shown to me by others? I would love to, but I don't. Unless I have been out in the field with someone, so I can take their measure, I only share information to the highest profile locations.

This is frustrating for everyone. Sometimes folks walk away feeling offended or even insulted. I am sorry, but I am not making a judgement about you (and I am pointing my finger right at you); instead, I am making a judgement about YOU (now, I'm waving my arms in the general direction of humanity). I know that there are some people out there who cannot be trusted.

I have been accused of elitism or of selfishness. I am willing to take on that mantle, if it helps protect the sites and their plants.

2) Be careful of what hard data you share

Finding carnivorous plant sites can be very easy. There are many places that are set up with boardwalks, visitor centers, and other facilities making the sites perfect for visitation. Information about these can be located within five minutes by anyone with access to the web—I even list some on the FAQ on my *Sarracenia.com* domain.

In contrast, finding the more obscure places can take a great deal of effort. Indeed, locating carnivorous plant sites in the western USA and documenting them is one of my primary research efforts. It is huge fun and incredibly satisfying.



Figure 3: A *Sarracenia flava* poaching hole (Onslow County, North Carolina). Photo: Barry Rice.

As part of my work, I document my results and quietly store them in appropriate scientific data repositories for future botanists. But I enjoy this work so much I want to tell others about what I have seen! So, I often make videos of the sites, which I post online. I am no Stanley Kubrick, so my videos are remedial at best, but I hope that my (tiny) audience enjoys them. I also post my images on my web domain, and also on CalPhotos (calphotos.berkeley.edu). But note—I never post location information with my photographs or videos. At most I give the site’s state and county (or parish—yes, Louisiana, I’m looking at you!)—nothing more.

Folks—please do not provide detailed information in your online postings. Yes, your friends are reading your posts, but so are the damned shadowy ghouls who travel with baggies and shovels.

3) Avoid unintended disclosures in group scientific activities

Group scientific endeavors can be very useful—the Audubon Society has been conducting “Christmas Bird Counts” since 1900, and the resulting data products are no doubt valuable reservoirs of information. More recently, other community-fed data products are becoming popular, probably most notably iNaturalist. This app allows field users to upload information about any organism they encounter in the field. Unfortunately, this tool has great potential for poachers. Indeed, Tyler Gramley (plant administrator with the North American *Sarracenia* Conservancy)—who has logged many hours looking for carnivorous plants in the field—believes firmly that this app has directly contributed to poaching at many sites, “When an observation on iNaturalist is left unobscured for any period of time, the site will have visible signs of visitation and damage....All of which occur in the short timeline of the day, or within one day, in the wake of an observation.” While the culpability of this one app cannot be unquestionably proven, its users should show common sense. There are features on the app that allow the user to blur the precision by which the detailed location information is being reported.

As I write this, I am discussing these issues with staff at iNaturalist. I would like to set the defaults of most carnivorous plants in the USA and Canada to always be blurred. At least, this is my goal—I do not know if they will be interested in complying.

4) Avoid unintended disclosures by image metadata

Handheld electronics (phones, etc.) gather GPS data whenever you take a photo, and encode this GPS information in the metadata for the images. This can be useful when organizing your image sets. However, it can also be transmitted to other recipients if you send them the images. If you have an iPhone, you can change the default setting so GPS metadata is not stored on your images. As I write this, the instructions are simple:

- Open the Settings app on your iPhone;
- Go to the Privacy setting;
- Tap Location Services, then find Camera in the list of apps;
- On the options associated with Camera, select the NEVER or OFF setting.

Androids and other phones have comparable options. Research them and implement them. Now.

During the field trips that were part of the 2018 ICPS Conference in California, before we all ventured out into the field, Gina Morimoto coached all of us on how to turn off the location-encoding on our phones. Everyone was appreciative and understanding of this protocol.

I have experimented with transmitting location-encoded images, and have found that when I upload images to Facebook or transmit images via Facebook Messenger, the GPS data are stripped off. Meanwhile, if I text an image to someone, the GPS data are retained. I do not know if these results are universal—the best approach is to control the Location Services on your phone directly.

5) Finally, be a snitch (within reason)

Technology is constantly changing. We have not fully adapted to our information revolution. As such, well-meaning people are doing foolish things. If you are associated with an information-providing agency such as an herbarium, resist the urge to upload all the data you can to the Internet. Sensitive data, even for species without state or federal listing status, should be blurred before being uploaded.

If you see someone else uploading data that should be removed contact the data source immediately.

On the other hand, if you see anyone poaching carnivorous plants, I encourage you to proceed extremely carefully if you contemplate confronting them. I have been told by agency staff in North Carolina that some *Dionaea* poachers are heavily armed, and I have been advised—if I encounter poachers—to not attempt to engage with them or even photograph them or their vehicles. They are potentially highly dangerous.

If you have gone through all the hard work to sleuth, deduce, and locate a wonderful site—and have the scratches, muddy boots, and torn pants to prove it—good for you. Now work hard to make sure your experience does not come at the expense of the plants you love so much. There will always be poachers out there. I have resigned myself to that. So, ensure that the wonders you experienced by visiting an exquisite plant site do not result in a biological crime by someone else. Try to live in such a way so that the plants and sites you visited, benefitted by your presence.

So maybe, don't share all the knowledge you have. Show discretion.

Sorry, Leo.

The author would like to thank the vigorous, dedicated, and active online community of botanists and naturalists who are working so hard to protect these plants. Tyler Gramley has been invaluable in giving me perspective on some of the risks involved with data-sharing.

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GROW VENUS FLYTRAPS INDOORS

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Keywords: cultivation, Venus flytrap, *Dionaea muscipula*.

When old timer carnivorous plant geeks get together, the conversation invariably gets to how many Venus flytraps (VFTs) they have killed. Fifty years ago, we had an excuse. Part of that excuse is we had no clue how to grow them. There were no books about growing carnivores. But even when books were eventually written and Carnivorous Plant Newsletter came into existence, the information was not exactly correct for growing VFTs indoors. The authors grew their plants in greenhouses and described what happens in greenhouses. I will not bother giving references here because it would be every carnivorous plant book with cultivation information that I have seen, except, I will give Adrian Slack (1979) kudos for not saying Venus flytraps will die if they do not get winter dormancy.

I am guilty of providing misinformation as well and I am sure there are still vestiges of dormancy for indoor VFT plants on the ICPS website that I have yet to purge. This does not change what you do for outdoor plants. If you grow your VFTs outside, you do have to pay attention to climate and dormancy. If your plants are outside and the temperature is going to be cold enough that the growth point of the plants will freeze, they **MUST** be dormant when it freezes, or you must put them in a sheltered location. I lost a lot of VFTs from a late freeze about 10 years ago. They survived being frozen solid twice during the winter but had started growing when we had a late spring freeze that even damaged native plants. I decided at that point I was done growing VFTs outside. Besides, when did I see them outside? I have to live with my plants. That means growing VFTs indoors where I can see them all the time.

I do not claim that how I grow my VFTs indoors is the best way ever or even if someone tries to duplicate what I do that there will not be problems. So much depends on the exact details of what you can and cannot do. If what you are doing works for you, great! Some of the details are general for any plant you grow in pots; others are critical for VFTs.

VFTs do not need high humidity. In the house I use terrariums I made that are leaky and never fog up. The point is to see the plants, not just to have them. Without the terrariums I would have to refill the water trays every other day because our humidity is very low. I do grow a lot of VFTs in my garage under lights without terrariums and have to refill the trays daily during hot weather. I cannot tell any difference between plants grown with and without terrariums.

The size of the pot and the soil mix have to be matched to the needs of the plants. VFTs are not bog plants that can be grown long term semi-aquatically or in saturated soil. But they do need to be in a wet soil typical for fen plants. Fen plants are easy to grow in greenhouses and outside because they like lighter soils and to be hosed off regularly. They are not so easy in houseplant situations. VFTs themselves do not use much water. They have few roots that are essentially drinking straws so are not very efficient extracting water from damp soil. You need to balance available water for the plants and the fact that old leaf bases that make up the bulb will rot in soggy soil. You want to delay bulb rot as long as possible.

I use 8 cm square by 9 cm tall pots because they fit perfectly in standard “1020” plant trays. It is difficult to get pots of this size now. There is no reason not to use bigger pots with multiple plants per pot or whatever else you can find in the way of tall pots or plastic cups. Larger pots will also help maintain enough soil moisture if for some reason the trays or water dishes dry out for a while.

Pots that are 10 cm square and 12 cm tall are easy to get and work great for multiple plants. They just do not fit nicely in 1020 trays.

Pick a soil mix that keeps the roots fairly wet but not so wet as to become anaerobic. I use 50:50 peat:sand. What exactly does “50:50” mean? Half and half by volume? By weight? I have no clue. I say that to mean “not overly sandy” and “not overly peaty”. Pure peat tends to become stinky. Pure sand would not wick up enough water. You want a nice balance of the two. I use more sand in shallower pots and more peat in deeper pots.

I only use a particular brand of peat that is a little hard to find, from a company that specializes in potting soil. The most common brand of peat is usually much too salty. The peat is mined as an inexpensive garden soil amendment and is not the quality you would like for a potting mix. I also wash/rinse the peat before I use it.

The sand I use is relatively coarse, graded, and washed. It is available in various mesh sizes where the “mesh” is the number of wires per inch in the sieves. I have found 14 and 16 mesh silica sand to be best but have had to settle with 12 and 20 mesh when that was all I could get. It is usually only available at specialized construction materials companies. Finer sand such as filter sand can pack tightly and keep water from wicking up through the soil mix. If all else fails, you might try sieving builders sand to remove the finest particles.

I never use perlite, pumice, vermiculite, or regular potting mix for my VFTs. Perlite and pumice are generally too salty, and many brands add fertilizer. Vermiculite breaks down into mush quite quickly. Regular potting mix is generally too coarse from the ground bark and leaf compost, has been “corrected” to a neutral pH, and has added fertilizer. I do not use long fibered *Sphagnum* for my VFTs because I think it should only be used for plants where it is the only good option. However, it may be the only option for some growers, and it does work great as a peat substitute.

I believe it is critical that most of the VFT bulb not be in the wet soil mix. I plant my plants with the bottom of the bulb and the roots in the soil mix, then top up the pot with about 2 cm of pure sand (Back Cover). The pure sand serves a number of purposes. The sand helps keep the older part of the bulb from rotting. With the sand, you do not get a mat of moss that the new growth has to plow through, and the moss could cause the soil mix to become anerobic. The sand allows air to get to the deeper soil. The sand also is not attractive to fungus gnats. Fungus gnat larvae are a bane of small plants and I do not want them in my terrariums. As the plants grow, they tend to find the level they like. I find they do not dig deeper into the soil. The bulb remains in the sand layer. The only problem with using dry sand in pots in the house is if the pots tip over, they can make a big mess.

The soil in the pots must never dry out. Some of my indoor VFTs constantly sit in 1 to 2 cm of water. Others get water added to the trays up to 3 cm deep when the tray dries out. I never top water them except during the repotting process. I am fortunate that my city tap water is usually around 50 ppm of dissolved solids. Most people probably have to invest in RO water or collect rain water. Bottled drinking water and filtered water can be deadly.

The plants need at least 15,000 Lux of white LED light for about 14 hours a day. The plants will grow even better with 25,000 Lux. This much light requires 20 to 30 W of white LEDs per 30 cm diameter of growing area. In the house I only use 3000K, 90+ color rendering index LEDs because of the enhanced deep red to better match the chlorophyll absorption spectrum. To me the plants look best under these lights as well. This is a lot of light. The plants need that much light to grow well. It is hard to get that much light from fluorescent lights without having heat issues. In the garage I use a mix of white LEDs and purple LED plant lights. You need the white light because you cannot see the purple light very well in spite of the fact it is very intense. Because of different designs of purple plant lights, the ratio of blue, red, and white light, and the difficulty measuring the

amount of light, I do not know what to recommend. Also, you have to be careful of manufacturers advertising “watt-equivalents” instead of actual watts.

It is critical that the plants be fed regularly. Strong light and regular feeding are the keys to growing VFTs indoors long term. In other words, the plants must be growing constantly and to do that they need lots of light and lots of food. I feed the plants with rehydrated freeze-dried bloodworms, which is a kind of fish food. The more you feed the plants, the faster they grow and larger they get. If their growth slows down, feed them. (See Figure 1 and the ICPS website has a page on feeding plants with bloodworms: <https://www.carnivorousplants.org/grow/feed/bloodworms>).

I do not know what temperature triggers dormancy in VFTs. My plants in the house do not slow down during the winter. They get 15 hours of LED light daily but do tend to bloom in the spring, presumably because they get some sunlight before the LEDs turn on at 7 AM. The plants in the garage slow down during the winter but never go dormant. Their lights are on a timer that adjusts for natural sunrise and sunset. I just keep feeding them to keep them growing because they are plants I am propagating.

The plants need to be repotted every year or two. At some point the plants will have grown across the pot and slammed into an edge. It is also possible for the soil to get too salty or something else bad to happen so the roots die and the new traps turn black before they open. Repot the plant in fresh soil as soon as possible if the plant will not eat. Otherwise feed it well, then after a few weeks



Figure 1: Feeding a Venus flytrap with rehydrated freeze-dried bloodworms.



Figure 2: (A) A Venus flytrap that has been growing indoors and is in need of repotting because it is starting to hit the side of the pot. (B) The plant unpot, trimmed, and older part of the bulb cut off. Notice how few roots there are. You use large pots to retain water, not because the plant can become root-bound. (C) The division planted in peat/sand soil. Note almost all of the plant is above the peat/sand soil level. (D) The division back with its buddies in the terrarium after the pot has been topped up with pure sand.

repot it. Do not reuse the soil; it can be used in the garden or for outdoor-only carnivores that get rained on. (See Figure 2)

When you repot the plants, you will need to reduce the size of the bulb. Use this opportunity to increase your collection of plants or to have plants to give away. (See Figure 3 and there is a page on the ICPS website about using the old bulb leaf bases to produce new plants: <https://www.carnivorousplants.org/grow/propagation/DionaeaLeafPullings>).

In this day and age there is no excuse not to grow Venus flytraps long term indoors. No refrigerator is necessary. Just keep the plants growing year-round. Doing so was somewhat problematic before LED lighting. Now there are lots of effective lighting options. If you cannot collect enough



Figure 3: (A) Leaf base pullings from the old part of bulb planted in soil before 1 cm of sand is added to the pot, completely covering the leaf bases in sand. (B) After six to 12 months, the leaf pullings should look like this.

rain water when you need it, RO water is generally available. And high protein fish food such as freeze-dried bloodworms is more available now than in the past. The hard part can be finding decent soil ingredients. In the end your plants can live with you instead of just lurking somewhere you only see them occasionally.

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QUEST FOR THE ORIGIN OF *HELIAMPHORA HETERODOXA*.
REPORT OF MAY 2018 EXPEDITION TO VENEZUELA WITH
CARNIVOROUS FLORA OCCURRENCES IN THE BASE AREAS OF PTARI-TEPUI

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Introduction

In May 2018 I had the opportunity to co-organize an independent expedition to the areas surrounding the massif of Ptari-tepui and Sororopan-tepui in Estado Bolívar, Venezuela. My group led by a team of Pemón guides from the community of Santa Teresita de Kavanayen succeeded in reaching the location of the first scientific expeditions to the massif of Ptari-tepui led by W. Phelps in February 1944 and J. A. Steyermark in November 1944, including re-discovering the locus classicus of *Heliamphora heterodoxa* which had not been accurately located ever since the original publication of the taxon in 1951 (McPherson *et al.* 2011).

Report of the expedition

The expedition started in the town of Santa Elena de Uairen in Estado Bolívar, Venezuela where our group assembled on May 2nd, 2018. The following day on May 3rd we drove north on the Troncal 10 Road, carried out observations of *Heliamphora* and other carnivorous plant populations in the savannas by the roadside, around the marshlands in close vicinities of Rio Apongua, and then continued southwest to the Pemón village of Santa Teresita de Kavanayen. Along the way we made short stops to visit the most popular touristic highlights of the Gran Sabana, such as the Jasper Creek (*Quebrada de Jaspe*), Salto Kama Meru, and the famous tepui viewpoint, *Mirador el Oso*. After reaching Kavanayen a short meeting was organized with the leader of the village and members of the Parupa scientific station and plans were discussed for the upcoming days.

In the following morning on May 4th the team, led by guides from Kavanayen, advanced westwards and started the approach towards the southwestern slopes of Ptari-tepui. The first day of trekking was spent on crossing the savanna to a lower forest campsite called Kavanaru (being an Amerindian name of the cock-of-the-rock bird, *Rupicola rupicola*). The savanna had a notable presence of carnivorous flora typical to the Gran Sabana region, including populations of *Drosera felix*, *Drosera roraimae*, *Catopsis berteroniana*, *Utricularia subulata*, *Utricularia hispida*, and *Brocchinia reducta*. In addition, *Utricularia olivacea*, *Drosera kaieteurensis*, and *Genlisea* sp. plants were found around a rest spot by a waterfall called Iwarakaru Meru approximately halfway through the trail (Fig. 1). Within the perimeter of camp Kavanaru, several *Utricularia jamesoniana* plants were spotted growing on mossy tree trunks.

The objective on May 5th was to reach Punto Phelps – a small cave campsite on the southwestern foothill of Ptari-tepui, named after the American ornithologist William H. Phelps who was the first to lead a scientific expedition in that area in February 1944 (Zimmer & Phelps 1944). The trail became significantly steeper and more difficult. Before reaching the campsite, the group traversed an elevated plateau area connecting the slopes of Ptari-tepui and Sororopan-tepui and passed by a number of scattered patches of typical, highland moist savanna vegetation divided by *Clusia* sp. and *Bonnetia sessilis* shrublands and savanna-forest mosaic vegetation (Fig. 2). This particular location



Figure 1: Highlights of the trail to camp Kavanaru: 1) *Catopsis berteroniana*; 2) View over Ptari-tepui from Iwarakaru Meru; 3) *Drosera kaieteurensis*; 4) *Utricularia olivacea*.

reminded of the areas where some of *Heliamphora heterodoxa* collections had been made in November 1944 and described by J. A. Steyermark in his 1951 *Fieldiana* record (Steyermark 1951). No *Heliamphora* plants were spotted, yet the climb provided the first encounters with members of flora typical to higher elevations in the Pantepui – plants such as *Brocchinia acuminata*, *Orectanthe scepterum*, or *Stegolepis ptaritepuiensis*.

A particularly interesting area was found around a rest spot by the Iwore Meru waterfall, where the vegetation became a lower cloud forest characterized by the presence of a significant number of Bromeliads, Orchids, Aroids, and members of the *Rapateaceae* family, such as *Saxofridericia regalis*.

After reaching Punto Phelps, a memorial plaque was found left by W. Phelps inside the cave campsite commemorating his visit to the spot in February 1944. I noticed an inscription on the plaque made probably with a nail or some other impro-

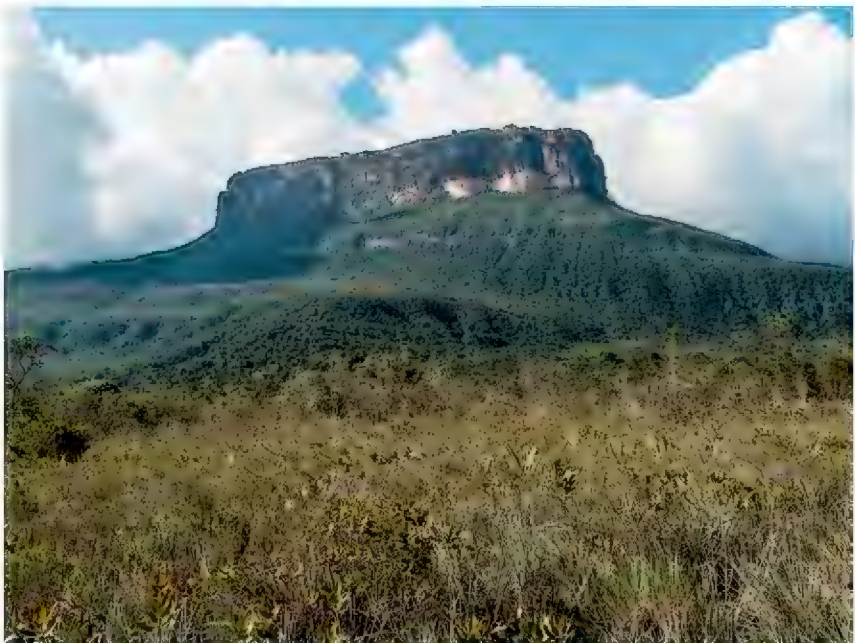


Figure 2: Ptari-tepui seen from one of the savanna patches on the plateau area interlinking to the slopes of Sororopan-tepui.



Figure 3: Highlights of the upper part of the Ptari-tepui trail: 1) Southwestern wall of Ptari-tepui seen from Punto Phelps camp; 2) Memorial plaque left in the cave at Punto Phelps camp, with J. A. Steyermark’s “vandalized” inscription; 3) Sandstone wall habitat with *H. collina*; 4) *H. collina* population growing on the rock wall.

vised tool, saying: “Julian Steyermark, 10 Noviembre 1944”. That inscription served as a clear evidence that the area was indeed the location of Steyermark’s original *Heliamphora heterodoxa* collections as it corresponded to the dates put in the herbarium specimens (Fig. 3).

May 6th was spent on a strenuous climb of the southwestern slope of Ptari-tepui with the objective of reaching as high as possible, making observations focused on carnivorous plant populations encountered along the way. The trail became increasingly steep, muddy and difficult to cross. Several parts required nearly vertical climbing using tree branches and roots to get around steep rocky surfaces and narrow, muddy ridges. When an exposed portion of the sandstone wall was reached at approx. 1800 m a.s.l., a large population of *Heliamphora collina* was found growing attached to the rock wall accompanied by *Utricularia alpina* and *Drosera roraimae* plants (Fig. 3). This observation confirmed that *Heliamphora collina* is indeed present on the cliffs of Ptari-tepui, as it had been suspected by some other researchers (McPherson *et al.* 2011).



Figure 4: *Heliamphora collina* found among shrub vegetation on the slopes of Ptari-tepui. Note the close morphological similarity to *Heliamphora folliculata* leaves.

Advancing further up the trail several other species of carnivorous plants were encountered, namely *Drosera arenicola*, *Utricularia quelchii*, *Utricularia amethystina*, and *Brocchinia reducta*. More high-tepui species were becoming present, such as *Maguireothamnus speciosus*, *Brocchinia tatei*, *Brocchinia steyermarkii*, *Pterozonium* ferns, *Elaphoglossum wurdackii*, *Ledothamnus* sp. or *Connellia* sp. More *Heliamphora collina* plants could be found growing around small patches of vegetation on exposed rock faces and around shrubs (Fig. 4).

The trail ended on the surface of one of the large sandstone boulders which were common in the slope areas. The summit of the boulder was partially bare rock, and in some parts thick shrub vegetation growing all around the slopes (Fig. 5), which directly matched the description of the location left by J. A. Steyermark on his *Heliamphora heterodoxa* type collections: “matted on mossy exposed top of big boulder; (...), Ptari-tepui, *Bonnetia roraimae* forest on southwest-facing shoulder, altitude 2000-2200 meters” (Steyermark 1951). Among the shrubs a significant amount of *Heliamphora* plants were found, which after closer observations were identified as a hybrid swarm of two species, *Heliamphora collina* and *Heliamphora purpurascens*.

Due to deteriorating weather conditions the group retreated back to Punto Phelps after spending less than an hour on the top of the boulder and continued down to Kavanaru camp the same day.

The expedition to Ptari-tepui slopes ended on May 7th when the group returned from camp Kavanaru to Kavanayen. Following days were dedicated to exploring various locations in the Gran Sa-



Figure 5: Vegetation on the top of the sandstone boulder location: 1) *Bonnetia roraimae* shrub vegetation with *Stegolepis*, with Mona-tepui visible in the background; 2) Sandstone boulders on the southwestern talus slope of Ptari-tepui, looking SE; 3) Shrub vegetation on the boulder, looking NW; 4) *Heliamphora collina* x *purpurascens* growing among shrubs on the top of the boulder.

bana, in the vicinities of Kavanayen and Parupa scientific station. Various locations were visited, for instance forests and savannas around Salto Apongua and Toron Meru. The expedition concluded with a trekking from Uroy-Uaray to the community of Wuarapata, near the base of Tramen-tepui.

Acknowledgements: Members of the community of Santa Teresita de Kavanayen in Estado Bolívar, Venezuela, especially Yoeli, Carlos, and Rosaura Chani are thanked for accompanying and providing guidance during the fieldwork that was carried out by the author on Ptari-tepui and its vicinities. Izabela Stachowicz is thanked for co-organizing the expedition. Paolo and Mauro Costa are thanked for providing useful insights about the areas around Kavanayen and Ptari-tepui. Andreas Fleischmann is thanked for helpful comments.

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NEW CULTIVARS

Keywords: cultivar, *Dionaea* ‘FFT Stegosaurus’, *Dionaea* ‘Génépine’, *Dionaea* ‘Morano’, *Dionaea* ‘Axe’.

Dionaea ‘FFT Stegosaurus’

Submitted: 7 July 2019

Dionaea ‘FFT Stegosaurus’ (*Dionaea* ‘FFT Estegosaurio’ in Spanish) differs from other *Dionaea* sawtooth-type cultivars by its teeth pattern; the cilia consist of one long tooth in between two smaller teeth. Traps are green, but turn light red when exposed to direct sunlight. The foliage is lime green, the leaf petioles do not become thin during the summer, and maintain part of the leaf blade. The foliage does not grow upright during the summer, but remains relatively close to the ground as other low growing cultivars. FFT stands for the name of my nursery, “Fayetteville Fly Traps”.



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Dionaea ‘Génépine’

Submitted: 1 September 2019

Dionaea ‘Génépine’ was selected from seedlings germinated in 2015. The plant is green with many white patches and beautiful white spines.

The name Génépine is a combination of Génépi and épines. The plant reminds me of the traditional green herbal liqueur, Génépi, on the snow. Génépi is based on the *Artemisia* plant which grows only in the mountains (2500-3000 m), where snow remains for a long time. The French word for spines is épines.

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Dionaea ‘Morano’

Submitted: 19 September 2019

I selected *Dionaea* ‘Morano’ in 2016 from seedlings germinated in 2013. The unique characteristics were evident in the young plants. *Dionaea* ‘Morano’ produces prostrate leaves. The leaves are about 6 cm long and the traps reach a maximum length of 15-16 mm. The traps are without teeth (only the first traps at the beginning of the season have a very short set of teeth) with a bright red internal coloring and forming a thin and very evident white “lunette” when they are adults. There is a marked contrast between the bright green of the petioles and the red of the traps.

Dionaea “Morano” must be reproduced vegetatively by rhizome or leaf/floral scape cuttings to preserve the unique characteristics of the cultivar.



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Dionaea 'Axe'

Submitted: 19 September 2019

I selected *Dionaea* 'Axe' in 2016 from seedlings germinated in 2013. The unique characteristics were evident in the young plants. Early-season petioles have an erect habit and present the first traps with very small teeth that will be absent in the following ones and will give the plant the appearance of an axe, hence the name. The adult leaves are about 6 cm long and the traps have a slender shape, they narrow in the farthest part of the petiole and reach 16-17 mm. The formation of a red line on the edge of adult traps in the area at the base of the teeth will give the axes of this unique specimen an even more bloody appearance. Towards the end of the season, the last traps will be prostrate with the formation of teeth and a red/orange color in the surface of the inner trap surfaces.

Dionaea 'Axe' must be reproduced vegetatively by rhizome or leaf/floral scape cuttings to preserve the unique characteristics of the cultivar.

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